Beyond the ocean: Contamination of freshwater ecosystems with (micro-) plastic particles

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Environmental context

Microplastics in freshwater ecosystems are an increasing issue, with the few available studies suggesting high contamination worldwide. Reliable data on concentrations, fluxes and polymer types in continental aquatic environments, including urban water systems, are needed. High-risk polymers and associated or adsorbed chemicals have to be identified, as well as their effects on both organisms and ecosystems. Therefore, numerous challenges arise to assess possible adverse effects.

Abstract

Massive accumulation of plastic particles has been reported for marine ecosystems around the world, posing a risk to the biota. Freshwater ecosystems have received less attention despite the majority of plastic litter being produced onshore and introduced into marine environments by rivers. Some studies report not only the presence of microplastics in freshwater ecosystems, but show that contamination is as severe as in the oceans. In continental waters microplastics have been observed in both sediments (predominantly lakeshores but also riverbanks) and water samples (predominantly surface water of lakes and rivers). This review highlights recent findings and discusses open questions, focusing on the methodology of assessing this contaminant in freshwater ecosystems. In this context, method harmonization is needed in order to obtain comparable data from different environmental compartments and sites. This includes sampling strategies (at spatial and temporal scales), sample treatment (taking into consideration high levels of organic matter and suspended solids) and reliable analytical methods to identify microplastics.

Keywords:

plastic debris, microplastics, freshwater ecosystems, emerging contaminants, plastic separation,

polymer identification

1. Introduction

Artificial polymers are lightweight, durable, display excellent thermal and electrical insulation properties and can be formed into almost any shape. These characteristics render them suitable for a huge variety of applications in almost every sector of our everyday life. Consequently, the worldwide production of plastic has increased from 1.5 million tons in 1950 to 288 million tons in 2012 [1]. A large proportion of plastic is used by the packaging industry for solely disposable use [1]. Given the extensive use of these materials, post-consumer plastic waste has dramatically increased while the percent recycled remains low. For instance, only 26.3% of all plastic waste in Europe was recycled in 2012 [1]. Plastic waste can enter the environment, for example, from poorly managed landfills or by carelessly discarded post-consumer products. Since plastic debris can often be transported by wind or direct runoff after rain events, a large proportion of this waste inevitably reaches aquatic ecosystems where it then accumulates. This contamination not only includes plastic debris characterized by a large size but also so called microplastics. This term was first used in 2004 to describe very small fragments of plastic observed in sea samples (20 μm in diameter) [2]. The definition has since been broadened to include all particles < 5 mm [3, 4], although a subdivision into large (L-MPP: 1-5 mm) and small microplastic particles (S-MPP: 1 μm-1 mm) has been introduced by several authors (e.g. [5-7]). Microplastics can be further classified into two kinds based on their origin. Primary microplastics are specifically engineered for various applications such as personal care products or can be in the form of pre-production pellets. So-called secondary microplastics result from degradation of macroplastics caused by UV radiation, mechanical abrasion, biological degradation and disintegration [8]. This seems to be a continuous process, most likely leading to very small particles on even the nanoscale [9, 10]. Synthetic clothing can be regarded either as a source of

45 primary or secondary microplastic fibers.

46 Detectable amounts of small plastic debris were documented in the open ocean as early as 1972.
47 Neuston net samples of surface plastic concentrations in the Sargasso Sea contained an average of
48 3,500 pieces/km^2 [11]. Concurrent studies reported plastic debris in Western Atlantic and North
49 Pacific surface waters [12-14] , although distribution was extremely variable. From the 1960's to the
50 2000's, many studies using direct surface debris measurements or seabird ingestion as a proxy
51 reported the massive occurrence of microplastic in marine environments around the world [2, 15, 16].

52 Both primary and secondary microplastics can enter the continental aquatic environment through
53 several pathways (Figure 1). One of the main sources is inadequate end-of-life treatment of plastic
54 debris. This debris enters aquatic systems directly by water run-off or via stormwater and wastewater
55 treatment plant (WWTP) outlets.

56 Additionally, granulated polyethylene (PE), polypropylene (PP) or polystyrene (PS) particles, used
57 for example in skin cleaners, can be introduced into wastewater [17]. Furthermore, it has been shown
58 that laundry washing machines discharge a large amount of plastic fibers into wastewater, with one
59 study estimating that a single wash can produce 1,900 fibers [18]. Industrial activities also contribute
60 to the amount of microplastics in freshwater/aquatic ecosystems. High amounts of microplastic
61 particles and fibers have been detected in the vicinity of industrial plants involved in paper
62 production [19]. Synthetic fibers are also known to contaminate sewage sludge [20]. This observation
63 suggests that WWTP at least reduce the amounts of synthetic fibers in sewage effluents. However,
64 the use of sewage sludge for agricultural fertilization can still contribute to environmental
65 microplastic contamination. Moreover, plastic mulching could be another terrestrial source of
66 microplastics but to our knowledge, these pathways have not yet been sufficiently documented [21].

67

68 Finally, atmospheric inputs cannot be ignored. Since plastic fragments are transported by the wind,
69 this must be also the case for microplastics. As for some organic micropollutants, atmospheric inputs

should be investigated since it could represent an important transfer vector [22].

Elucidating sources and pathways of microplastics in freshwater ecosystems will be a major challenge for future research. This information will be the basis for management strategies to tackle problems arising from this emerging environmental contaminant. In the light of available data, a specific regulation was adopted by the European Union in 2008. The Marine Strategy Framework Directive (2008/56/EC, MSFD) [23] aims to more effectively protect marine environments across Europe, aiming to achieve good environmental status for European marine waters by 2020. Among the MSFD qualitative descriptors for determining good environmental status, indicator 10 is related to marine litter properties and quantities, including criteria to assess trends in amount, distribution and, where possible, composition of microparticles with a focus on microplastics. Similarly to the Marine Strategy Framework Directive, the Water Framework Directive (2000/60/EC, WFD) [24] aims to achieve good chemical and ecological status of all water bodies, including rivers and lakes. Until now, contamination with plastic debris (from micro- to macroplastics) has not been considered. This gap could be explained by the lack of i) data related to the occurrence and associated effects of microplastic contamination in freshwater ecosystems, and ii) robust and accurate methodologies to assess concentrations of microplastics in freshwater (environment and biota). The aim of the present critical review is to summarize available data on microplastics in freshwater ecosystems, and to identify and discuss scientific challenges surrounding this issue.

2. Microplastics in freshwater environments

In contrast to the large amount of literature describing marine environment contamination with plastic waste, only a few studies have addressed the issue of microplastic contamination in lakes and rivers. Estuary microplastic abundance has also received little attention [25-27], but given the strong influence of salinity gradients and tidal movements in these systems, only freshwater ecosystems

were considered in this review.

2.1. Lakeshore and riverbank sediment samples

Microplastic occurrence in lakeshore and riverbank sediments

Several studies reported microplastic contamination of lakeshore and riverbank sediment samples from continental aquatic systems in Europe, both North and South America, and Asia. To our knowledge, no studies have focused on lake bottom sediments.

Reported levels of microplastics in lakeshore sediments vary by a factor of 1,000 across the reviewed studies (Figure 2). In lakeshore sediments of Lake Garda (Italy), the authors reported that the north shore contained 1,108 ± 983 microplastic particles/m^2 whereas only 108 ± 55 microplastic particles/m^2 were observed on the south shore [9]. Spatial distribution of microplastics suggested that wind, lake morphology and the consequent currents are responsible for the observed pattern. The study showed that the most abundant polymer (45.6%) was PS, while PE was still highly abundant (43.1%). Polyamide (PA) and polyvinylchloride (PVC) were also identified down to a size of 9 µm. This study highlighted that the particles observed were fragments originating from the breakdown of larger particles (most likely post-consumer products), given that the scanning electron microscopy analysis revealed distinct signs of degradation.

Another study assessed microplastic contamination of lakeshore sediments along Lake Geneva (Switzerland) [28]. The results of the study are indicated in particles per liter of sediments. Concentrations varied from 1 to 7 particles/L [9], the predominant polymer was PS. Microplastic concentrations in a second study of Lake Geneva lakeshore sediments [29] varied from 2,656.25 to 5,018.75 particles/m^2, far greater than the highest concentration reported in lakeshore sediments of Lake Garda. A predominance of textile fibers, representing more than 90% of identified microplastics, was observed in the latter study.

In North America the distribution of particles along the lakeshores of one of the Laurentian Great

117 Lakes (Lake Huron Canada, USA) has been studied [10]. In this work, particles were directly
118 collected at the shoreline, then separated into three groups: [< 5 mm plastic pellets], [> 5 mm broken
119 plastic fragments], [all size PS particles]. The predominant microplastic form on Lake Huron
120 lakeshores was dependent upon sample location. In one of the sampled sites, over 94% of observed
121 plastic particles were industrial pellets, while at a different site pellets made up only 15% with the
122 largest fraction consisting of PS foam. The majority of pellets, predominantly PE, were observed
123 proximal to an industrial sector.
124 In a second publication [30], the abundance of plastics on the lakeshores of Lake Huron, Lake Erie
125 and Lake St. Clair (Canada, USA) was assessed and compared with those previously determined for
126 Lake Huron. As in the first study, high numbers of pellets were reported in comparison to fragments
127 and PS foam. Especially at Lake Huron 92% out of 3,209 particles were industrial pellets. The same
128 was true for Lake Erie with 39% out of 1,576 particles. The abundance of industrial pellets was
129 highest next to industrial areas and decreased along the shoreline. Along Lake Erie, PA was one of
130 the main polymers observed, making up 33% of the total plastic and coming mainly in the form of
131 pellets (47% of all pellets). Compared to other studies, the observed abundance of plastic debris is
132 rather low (Lake Huron: 4.75 ± 11.83 particles/m², Lake Erie 1.54 ± 1.01 particles/m², Lake St. Clair
133 1.72 ± 2.64 particles/m²). This might be mainly due to restricted sampling of visible fragments and
134 pellets, allowing microplastics invisible to the naked eye to be overlooked. Nevertheless, the Great
135 Lakes display a high degree of contamination with plastic debris which is mainly due to industrial
136 pre-production pellets making up 66% of the plastic load.
137 In general, the lakeshores of Lake Huron, Lake Geneva and Lake Garda contain lower concentrations
138 than marine beaches known to be highly contaminated [4]. Lake Huron displays more ocean like
139 characteristics and contains a similar concentration of plastic pellets as marine systems [31]; this is in
140 contrast to Lake Garda or Lake Geneva where only low concentrations of pellets were reported.
141 Local conditions/sources might explain these differences.

142 Studies of microplastics in river sediments are rare. A sediment study of St. Lawrence River
143 (Canada) showed high microbead abundance (similar to those used in consumer products) with sizes
144 between 0.5 and 2 mm [32]. Although the presence of microbeads was ubiquitous in all sediment
145 samples (mean 13,832 particles/m^2), some sites presented much higher concentrations (maximum
146 10^5 particles/m^2). This spatial distribution is potentially explained by environmental factors affecting
147 sedimentation.
148 One single study investigated riverbank abundance and composition of macroplastics [> 1.5 cm]
149 from four rivers flowing into the south east pacific, starting at the headwaters down to the river
150 mouth [33]. Plastics were the prevailing litter items at most sampling sites, their number varying
151 between 15 to 73% of total collected litter items. Total abundance of plastics (including PS)
152 remained below 4 items/m^2. No specific pattern was observed along the river from headwaters to
153 river mouth. Thus hydrology did not seem to be a key factor determining plastics abundance; their
154 variability was more closely linked to land use and river shore accessibility, which facilitates such
155 activities as illegal waste dumping.

156 ***Sampling and separating microplastics from lakeshore and riverbank sediments***

157 At Lake Garda ‚sediment sampling was performed using random grid samples" [6]. The separation of
158 microplastics was performed using density separation with a solution of zinc chloride ($ZnCl_2$). To
159 analyze lakeshore sediment samples from Lake Geneva, two protocols have been applied [28, 29]. The
160 first consisted of direct collection of coarse plastic fragments at the lakeshore, but the authors didn't
161 specify the size of fragments collected. In the second method, sand samples were successively
162 separated using 5 and 2 mm sieves, after which water was added to collect floating particles.
163 At the sites on Lake Huron, Lake Erie and Lake St. Clair [10, 30], plastic fragments [< 10 cm] were
164 sampled from sandy lakeshores using stainless steel trowels. Sampling was performed at each
165 location using 1 m wide stripes running from the water to the vegetation line. These stripes were
166 placed along a 60 m transect parallel to the shoreline in 10 m intervals. Larger items were counted

at each site but were not collected. The few studies of river sediment microplastics used either different types of grab samplers taken at depths varying from 10 to 15 cm [32], or sampling was based on visual inspection within sampling circles at various locations within the river bed: i) river shore at the edge of the river, ii) mid bank between the river bank up to the high water mark and iii) upper bank outside the river bed [33].

Identification

Sampled plastic particles from lakeshore and stream sediments have been identified as polymers using a variety of methods. In three studies analysis was only performed by visual means [28, 29, 33]. Particle composition from the lakeshores of Lake Huron, Lake Erie and Lake St. Clair was determined with Fourier transform infrared spectroscopy (FT-IR) [10, 30]. Plastic particles observed in Lake Garda beach sediments have been analyzed using Raman microspectroscopy (RM) [34]. A rarely used method for plastic particle identification was employed in a study assessing the abundance of microbeads in St. Lawrence River beach sediments, where microbeads were analyzed using Differential Scanning Calorimetry [32].

2.2. Water samples

Microplastic occurrence in freshwater

Microplastic contamination of surface water has also been investigated, particularly in lakes. As seen in lakeshore sediment concentrations, those for lake surface water differ by a factor of 1,000 across different studies (Figure 2). In Lake Geneva (Switzerland) [28] authors reported an extrapolated density of 48,146 particles/km^2. Similarly, surface water of the Laurentian Great Lakes (i.e. Lake Huron, Lake Superior, Lake Erie) was sampled [7]. Plastic particles have been categorized in three groups: [0.355 mm – 0.999 mm], [1.00 mm – 4.75 mm], [> 4.75 mm]. Most plastic particles were observed in the smallest category, suggesting a similar risk for freshwater biota as reported for marine environments [8]. Several of the microplastic particles were green, blue and purple colored

191 spheres, likely stemming from facial cleaners and other personal care products. These were identified
192 as PE and PP. The mean concentration on the studied lakes was 43,157 particles/km^2, ranging from
193 zero to 28,0947 particles/km^2. Lake Erie alone accounted for 90% of the total plastics reported in all
194 three lakes and contained the two most contaminated areas. The fact that two samples were highly
195 contaminated compared to the rest of the samples is in concordance with other studies [9, 10, 30]. The
196 authors suggest that this high abundance results from converging currents, proximity to several coal
197 burning power plants and their downstream location from cities such as Detroit and Cleveland.
198 In a remote mountain Lake (Lake Hovsgol, Mongolia), an average density of 20,264 particles/km²
199 (997 – 44,435 particles/km², min-max values) was observed [35]. Particles were grouped into the same
200 three size classes as for Laurentian Great Lakes. Although Lake Hovsgol is a large lake with a
201 surface area similar to Lake Erie, its catchment is less densely populated. Despite this, contamination
202 with microplastic particles is significant, which the authors attribute to aerial transfer from distant
203 urban sources.
204 Concentrations of microplastics reported for rivers, is highly variable (up to a factor of 10^9; Figure
205 3), likely due to the different methodologies used. On the Greater London (Great Britain) rivers,
206 microplastic pollution was examined in an unpublished study of four sampling sites [36]. This work
207 reports a concentration between 3.3 and 9.9 particles/L in two sites. Two rivers in California, San
208 Gabriel and Los Angeles Rivers (USA), were also investigated [37]. Plastic particle numbers ranged
209 from 0.01 to 12.9 particles/L. After a rain event, smaller microplastics (1-4.75 mm) were 16 times
210 more abundant in the Los Angeles River than larger plastic particles (> 4.75 mm). Under the same
211 conditions in the San Gabriel River, small particles were only 3 times more abundant. In both rivers,
212 71% of the plastic items recovered were comprised of foam. Extrapolation of these results estimated
213 that 2.3 billion particles were introduced into the marine environment over a period of 3 days [37].
214 A study of the Danube River in Central Europe [38] revealed a mean (± standard deviation) plastic
215 abundance of 316.8 ± 4,664.6 items/1,000 m^3 (0.00032 ± 0.00465 particles/L) during a two year

survey (2010, 2012). The corresponding plastic input via the Danube into the Black Sea was estimated at 4.2 t/day. Industrial raw material (pellets, flakes and spherules) accounted for substantial portions (79.4%) of the plastic debris.

Concentrations of microplastics in the North Shore Channel in Chicago (USA) were investigated upstream and downstream of a WWTP outlet (Terrence J. O'Brien Water Reclamation Plan) [39]. A mean concentration of 1.94 particles/m^3 (0.00194 particles/L) was observed upstream of the WWTP while downstream it was 17.93 particles/m^3 (0.01793 particles/L). These results highlighted that WWTP discharges might represent an important primary source of microplastics in freshwater.

Two different mesh size nets were used to collect surface water on the Seine River (France) [22]. Using an 80 µm size net, concentrations of microplastics were reported to vary between 3 to 106 particles/m^3 (0.003 to 0.106 particles/L). In contrast, sampling using a 330 µm mesh size manta trawl yielded concentrations of only 0.28 to 0.45 particles/m^3 (0.00028 to 0.00045 particles/L). Most of the microplastics observed were fibers, and 52% of plastic fibers collected with the first method were smaller than 1,000 µm. Only 25% of plastic fibers collected with the second method were smaller than 1,000 µm.

In the Chesapeake Bay (USA), concentrations reached a mean of 246 g/km^2, corresponding to 260,000 particles/km^2, in one of the four estuarine rivers studied [40]. Concentrations demonstrated statistically significant positive correlations with population density and proportion of urban/suburban development within watersheds. It should be noted that the greatest microplastic concentrations also occurred at three of four sites shortly after major rain events.

Large drifting plastic debris were documented on the bottom of the Thames River (Great Britain) [41]. For this study, a total of 8,490 submerged plastic items were intercepted during a 3 month sampling period.

The abundance and composition of floating plastic debris along the Seine River (France) was also investigated [42]. This study focused on macroplastic pollution. A significant proportion of buoyant plastic debris consisted of food wrappers/containers and plastic cutlery. PP, PE and, to a lesser extent, polyethylene terephthalate (PET) were the most abundant types of polymers observed. A first extrapolation estimated that 27 tons of plastic debris are intercepted annually by a regional network of floating debris retention booms.

Sampling of surface water

In marine environments, manta trawl sampling is the primary method used to sample lake surface waters or more recently in rivers. In Lake Geneva, a 333 µm mesh manta trawl was utilized [28, 29]. Samples were then passed through a 5 mm sieve in order to separate macro- and microplastics. Other studies focusing on lake water contamination with microplastics also used a 333 µm net to collect samples [7, 35].

Manta trawls (333 µm) have also been used [22, 37, 39, 40] to sample river surface waters. Other types of nets/devices have been utilized in rivers: stream bed samplers and hand nets (sampling surface water < 1 mm mesh size) [37], eel fyke nets (sampling water next to the riverbed - no details on mesh size) [41], and stationary driftnets (sampling the top 0.5 m of the water column - 500 µm mesh size) [38]. One study combined the use of a manta trawl for particles down to 333 µm and a plankton net for particles down to 80 µm [22].

Organic matter removal

Although organic debris hampers the identification of plastic particles and especially microplastic particles, only two studies applied a treatment to the samples prior to analysis [35, 40]. One was focusing on lake water and the other on riverine waters. Both applied a wet oxidizing protocol with hydrogen peroxide in presence of an iron(II) catalyst to remove organic material from surface water samples.

263 ***Identification***

264 Plastic particles observed in surface samples from lakes and streams have been identified as
265 polymers in ways similar to particles detected in lakeshore sediments. Although the identification by
266 visual means is less reliable it was performed by 7 studies [22, 28, 29, 35, 37, 38, 41]. Spectrometric methods
267 have been used in 3 studies to identify polymers, namely IR-spectroscopy [26], FT-IR using the ATR
268 (Attenuated total reflection) technique [42] and RM [40].

269 **2.3. Biota samples**

270 Only two studies assessed freshwater biota ingestion of microplastic particles. The first was
271 performed in Lake Geneva (Switzerland). The gut content of 21 adult northern pikes (*Esox lucius*),
272 18 common roaches (*Rutilus rutilus*) and 2 common breams (*Abramis brama*) were analyzed but no
273 plastic fragments were found [28]. The second study assessed microplastics in the gut of wild
274 gudgeons (*Gobio gobio*) sampled in 11 French rivers [43]. For this purpose, fish guts were dissected
275 and subjected to direct visual inspection under a dissecting microscope. Microplastics, defined as
276 hard and colored fibers, were observed in fish from 8 rivers with an occurrence between 11 and 26%.
277 The authors showed that fish from urban rivers were more contaminated with microplastics
278 compared to those collected in rivers with low anthropogenic impact. This study confirms that
279 freshwater fish do ingest microplastics and supports further studies to characterize microplastic
280 contamination of river and lake biota. Both studies used visual inspection methods to identify the
281 plastic particles and fibers.

282 ## 3. Challenges to and recommendations for the analysis of microplastics in
283 ## freshwater environments – A crucial need for harmonization

284 One of the major challenges in microplastic research is the need for general definitions and method
285 harmonization. This will enable comparison of results between studies and sites, and should

encompass every step involved in the microplastic investigation, i.e. sampling, samples processing, identification and final statistics.

3.1 Definition of microplastics and units used

Microplastics comprise a heterogeneous assemblage of pieces that vary in size, shape, color, specific density and chemical composition. The definition of microplastic size varies in previous marine studies. This heterogeneity is also found in studies concerning the continental environment. To avoid this issue, our recommendation is to provide a common definition of microplastics which should support the establishment of a standardized sampling method and improve concordance between future studies. In the marine environment, studies consider microplastics as particles smaller than 5 mm in size [4, 44]. However, given possible uptake by different aquatic organisms as well as the handling during extraction and identification, we suggest a subdivision of the term microplastic in particles that can be optically identified with the naked eye as well as handled with tweezers and particles which are impossible to distinguish without optical tools and cannot be handled individually without optical tools (e.g. microscope, stereo microscope). This is in concordance with several authors of marine studies [4, 45] as well as limnetic studies [7, 9, 35]. Similarly the Technical work group defining suggestions for implementation of Monitoring of Marine Litter for the Marine Strategy Framework Directive suggested to separate between microplastic >1 mm and microplastic <1 mm due to the above mentioned reasons [46].

In addition to a different microplastic definition, studies often give their result in different units, making comparability almost impossible (Table 1). This is mainly due to different sampling, extraction and identification methods.

Studies assessing lakeshore sediments give particles per volume [28] or per sampled surface [9, 10]. This is comparable to marine studies, although the latter also provide measurements of particles per sediment weight [4].

For lake samples, concentrations are mentioned in particles per surface area [7], again comparable to marine surface water sampling [4]. For the river data particle abundance is generally reported as particles per water volume [36, 39]. It is possible sometimes to calculate from one unit to the other, but the required information must be provided. The usage of particles per biomass is not hepful in an environment with seasonal changes (e.g. algal bloom in spring and summer, clear water phase, zooplankton blooms).

In order to identify plastic sources and characteristics, studies should also categorize plastics into different shape/size classes. Separating fibers (1 dimension larger than the two other dimensions), fragments (2 dimensions are large in contrast to a small third dimension) and spherules (similiar extent of all 3 dimensions) would enhance comparability between different sites.

3.2 Sampling methods

Sediment sampling

Correct and representative sampling of an adequate matrix is the first step to assess environmental contamination. Unfortunately, the same methodological divergence seen in the marine system (for review see [4]) occurs in sampling methods used for lakeshore sediments. Sampling differs not only in the methodologies used and the volume sampled, but also in the sample location. In order to avoid local heterogeneities, we propose a combined sampling approach using sediment core samples (diameter 10 cm, depth 5 cm) taken along a 20 m transect (e.g. along the drift line at a lake system) at a distance of 2.5 m from one another, which would result in a sample volume of 4 - 6 liters. For rivers, a different methodology should be applied. Instead of running parallel to the waterline, the river bank could be divided into 3 short transects of 5 m stretched over the accumulation zone perpendicular to the waterline.

Water sampling

To date a comparison between results from different studies is nearly impossible because nets with

various mesh sizes are used. Moreover, river water is sampled from different positions both in sampling depth and distance from the riverbanks. The main challenges for method standardization are i) the spatial-temporal frame and ii) the utilized mesh size which controls the smallest particle size sampled. A mesh size of 300 - 333 µm is common in marine plastic and plankton research, and offers a tradeoff between good handling and accumulation of larger fragments resulting in a blockage of the net. Using a mesh size of 300 - 333 µm in the freshwater environment would enable comparison of data gathered from both marine and continental environments. However, the use of a manta trawl may lead to an underestimation of microplastics in a size range smaller than the mesh size [22]. In order to have a complete overview of plastic contamination, we suggest also using nets with a smaller mesh size or, when necessary, even bulk water sampling. This would be an important consideration given that especially the smallest particles present a greater risk to be ingested and subsequently translocated into an organism's tissues [47].

Sampling microplastics on the surface of inshore waters may present some technical difficulties compared to marine ecosystems. In contrast to rather nutrient poor marine ecosystems, the amount of organic matter is generally much higher, especially during periods of algal blooms or leaf fall in autumn. Additionally, during periods of high water levels, suspended matter, clay minerals and allochthonous inputs of organic material increase the risk of clogging nets. This might limit the sampling duration, and thus reducing considerably volumes sampled. Therefore, while towing a manta trawl seems optimal for marine water sampling, it might be necessary to modify the net configuration in order to sample lakes and streams.

In contrast to lakes, rivers present a permanent flow of water and the current velocity has to be considered in order to facilitate surface sampling. The current velocity can be either too slow (< 0.1 m/s) or far too high, the latter resulting in a high ram pressure hampering net inflow. River water sampling should be preferable performed from a fixed position. A second problem arises from the

need for a reference parameter to give either particles per surface or per volume. While in standing waters, length of the performed manta tow can be used to calculate either the volume or surface sampled, but in running water, determination of current velocity must be estimated using a flowmeter.

Additionally, in order to determine fluxes of microplastics in running water, it is important to identify both the spatial and temporal variability of plastic particles as well as their dynamics in the river (distribution along the water column, sedimentation).

3.3 Methods for sample processing

Separation of plastic polymers from inorganic/mineral material

If plastic particles are directly collected at the lakeshore or riverbank, it is very likely that especially microparticles will be overlooked. A crucial step in taking all plastic particles into account is therefore the extraction of microplastics from bulk environmental samples. Different density separation methods can be used to separate microplastics from sediments [4]. The used methodology can massively account for a large portion of uncertainty in the recovery success. Due to their surface properties, microplastics can attach to any surface they come in contact with. Thus, the amount of working steps should be reduced for density separation of microplastics. Since sample preparation and particle identification is time consuming, the procedure has to be optimized regarding the extraction method. A density separator was developed for this purpose. With the Munich Plastic Sediment Separator (MPSS), a sample volume of up to 6 L can be analyzed in one run [6]. It should be noted that for sediments including high amounts of organic particles or clay minerals, the sample volume should be reduced. For very small sample volumes (< 250 mL), a MPSS in smaller scale could be built. The MPSS offers a good recovery rate of microplastics, especially for S-MPP, and is commercially available [6]. However, other methods using elutriation followed by density separation [48] and fluidization, and finally followed again by density separation [49] were also efficient.

382 Protocols can be adopted to optimize microplastic recovery in different types of sediments (sandy or
383 clay soils, biota-rich sediments, etc.).
384 Depending on the density of the solution used for the separation, the range of recovered polymers is
385 highly different. Previous studies on marine or estuarine environments use mainly sodium chloride
386 solution (NaCl - 1.2 kg/L) [4, 25]. If all commercially produced polymers should be recovered, the
387 solutions used for future separations must have a density of at least 1.5 kg/L or higher [6, 50]. The use
388 of a higher density solution takes into account both potential additives that may increase the density
389 of the particles, and attached biota or organic particles. Recently suggested separation fluids are zinc
390 chloride ($ZnCl_2$ - 1.6 to 1.7 kg/L) [6] or natrium iodide (NaI - 1.6 kg/L) [48, 49]. Both are relatively
391 cheap but have the drawback of being moderately toxic for the biota. The separation fluid may be
392 used more than once, being easily recovered for example with the use of candle filters. A more
393 expensive method is the use of a non-toxic polytungstate solution that offers a density up to 2.0 kg/L
394 [10].

395 ***Removal of organic matter***

396 The separation of plastic particles from other organic materials (such as shell fragments, small
397 organisms, algae or sea grasses, and tar) has been shown to be necessary for marine matrices and, to
398 a greater extent, for continental environment matrices. These latter matrices contain more organic
399 material, especially in the case of eutrophic lake and streams. Performing solely density separation to
400 isolate plastic particles from sediment samples is not efficient in reducing natural organic debris.
401 Hence, treatment methods have to be applied to facilitate identification of plastic particles. Several
402 oxidation agents such as hydrogen peroxide (H_2O_2) [51], strong acids such as nitric acid (HNO_3) [52],
403 hydrochloric acid (HCl) and mixtures of sulfuric acid (H_2SO_4) with H_2O_2 [34] were applied to remove
404 organic material for limnetic but also for marine samples. However, methods utilizing strong acids
405 have to be avoided since they affect and degrade plastic polymers [48]. Therefore alternative anti-

406 organic treatments need to be developed. A wet oxidizing protocol was recently used in a variety of
407 studies, but the extent of degradation induced by the wet oxidizing protocol has not been analyzed
408 yet. Some polymers are affected by H_2O_2 at room temperature (e.g. PA & POM [53]).
409 Enzymatic digestion methods may prove to be more useful to remove organic matter without
410 affecting plastic polymers. In 2014, an enzymatic protocol for marine water samples was published
411 revealing that 97% of the organic material can be digested without doing harm to microplastic
412 particles [54]. The digestion step can be conducted on bulk samples or after the density separation of
413 sediment samples.

414 **3.4 Quality assessment/Quality control**

415 To achieve the development of a common protocol to sample, extract and identify microplastics in
416 freshwater ecosystems, QA/QC (Quality assessment/Quality control) approaches will be very useful
417 to ensure the quality of results, evaluate sources of variability and error, and increase confidence in
418 the data collected. During the sample processing, sampling and laboratory blanks following the same
419 analytical protocols should be performed. Caution should be exercised in wearing synthetic fiber
420 clothing and should be avoided during sampling, extraction and further processing through until
421 identification. Samples have to be covered in order to prevent airborne contamination. Plastic
422 extraction recoveries also need to be validated using, for example, sediments spiked with artificially
423 placed plastic fragments. The impact on artificial polymers of all organic matter removal methods
424 have to be examined. In addition, identification methods should be used that corresponds to the size
425 of the analyzed particles.

426 **3.5 Identification of polymers**

427 Counting and identification are crucial steps to address microplastic contamination. Visual
428 examination has been commonly applied to assess size and quantities of microplastics. However,
429 pure visual examination using light or electron microscopy cannot be used to reliably distinguish

between polymers and other particles or to determine the polymer type. This method may lead to an overestimation of the plastic polymer contamination. In one study, nearly 20% of particles less than 1 mm which were initially identified as microplastic by visual observation were later realized to be aluminum silicate from coal ash [7]. Hence, visual characterization and identification should be coupled systematically to a characterization technique. Reliable identification results are achieved with the use of spectrometric methods like FT-IR microspectroscopy and Raman microspectroscopy, or for some polymers by the use of SEM/EDS (scanning electron microscopy / Energy Dispersive X-Ray Spectrometer) [55]. Pyrolysis followed by GC-MS can also be applied [49, 56] , but information on particle shape/size is lost.

The visual identification of macroplastic for abundance estimates might be acceptable, especially for studies only sampling large fragments of obvious plastic products [33]. In this case, microscopic inspection of texture and surface characteristics can be accompanied by verification by a spectrometric method similar to that performed at Lake Erie and St. Clair [30]. L-MPP can be handled with tweezers and can therefore be easily placed under the ATR crystal of a FT-IR, put under a Raman or electron microscope, be inserted in Pyrolysis GC/MS, or prepared for Differential Scanning Calorimetry. S-MPP samples have to be captured on matrices (e.g. filters) and subsequently undergo either manual or automatized identification protocols using FT-IR or RM. Manual identification methods are highly time consuming.

3.6 Microplastic contamination impacts in freshwater

As documented in marine organisms [47], freshwater fish [43] and invertebrates [9], microplastics can be ingested by aquatic organisms. However, the effects induced by microplastics are poorly documented and major questions should be investigated to address this issue. Microplastic contamination of biota has been classically documented using gut contents. After ingestion, microplastics may be retained, excreted or translocated into other body tissues and fluids. Evidence

454 of translocation is available in rodents and humans where 150 µm particles of PVC and PS were
455 identified in the lymph and circulatory system [57, 58]. More recently, particles were proved to interact
456 with mammalian cells in the intestinal system [59]. A translocation experiment was performed with
457 mussels (*Mytilus edulis*) exposed to 3 and 9.6 µm particles. After 3 days, translocation to the
458 circulatory system was observed and consisted of a greater number of smaller particles compared to
459 larger microplastics [60]. In concordance with this, another study detected microplastic occurrence in
460 the soft tissues of cultured *Mytilus edulis* and *Crassostrea gigas* [52]. These studies confirm that
461 microplastics can be translocated into tissues, but further studies are needed to address this aspect in
462 freshwater organisms.
463 Ingested of translocated microplastics induce adverse effects according to their mode of action. The
464 effects can be categorized as follows:
465 i) mechanical impairments through swallowed plastics mistaken as food
466 ii) polymers and plastic associated chemicals (additives) may be endocrine-disrupting or toxic
467 iii) polymers can adsorb toxic organic pollutants, nanoparticles or metals which may evoke
468 adverse effects
469 iv) Finally, plastic debris may act as vector for alien species and diseases.
470 To improve our knowledge of the microplastic hazard, organic micropollutant contamination must be
471 investigated. Acting as passive samplers, microplastics can indeed adsorb some pollutants. To date
472 regarding to persistent organic pollutants, the contribution of microplastic to the total pollution in
473 freshwater remains unknown, although numerous studies on marine beaches exist [61]. Freshwater
474 systems present different conditions compared to marine systems (salinity, organic debris etc.), and
475 concentrations of persistent organic pollutants in the continental environment are expected to be
476 greater than in marine ecosystems. Hence, it is essential to address interactions between micro-
477 pollutants and plastics in freshwater, focusing specifically on urban lakes and rivers.

4. Conclusions

Although environmental contamination with microplastics is constantly in the media, almost no basic data on contamination in freshwater ecosystems exist. As summarized in this review, recent studies demonstrate an almost equal contamination as reported in the oceans. Therefore, it is of utmost importance to elucidate sources, fate, fluxes, and impact of microplastics and associated chemicals in freshwater ecosystems.

Sampling, separation and identification methods are key steps for an accurate characterization of microplastic contamination. However the methods described in the existing literature are very diverse and no common methodology has emerged. This is likely due to the novelty of the topic, and is not astonishing given that many of the studies were pilot projects. The imposed constraint, however, is the low comparability between studies using different methods. This methodological heterogeneity might be intrinsically more pronounced for freshwater (especially in rivers) due to many parameters related to the freshwater sampling (river flow, season, type of net, position of the net or manta trawl, water colon height, dynamic or static sampling, time of exposure, presence of suspended mater, vegetal debris, etc.). This could introduce various constraints leading to the use of different methods. The development of an improved, automated and harmonized methodology for detection and identification of microplastics appears a real challenge, but should regardless be a priority in order to improve comparability between future studies (in both continental and marine environments). Comparable data on contamination of different habitats are important for a reliable risk assessment, which will be needed for adequate mitigation and prevention measures in the future. Guidance for monitoring microplastics have been recently proposed to support the monitoring of microdebris in the marine environment within the Marine Strategy Framework Directive [62] but gaps have to be identified as preliminary steps.

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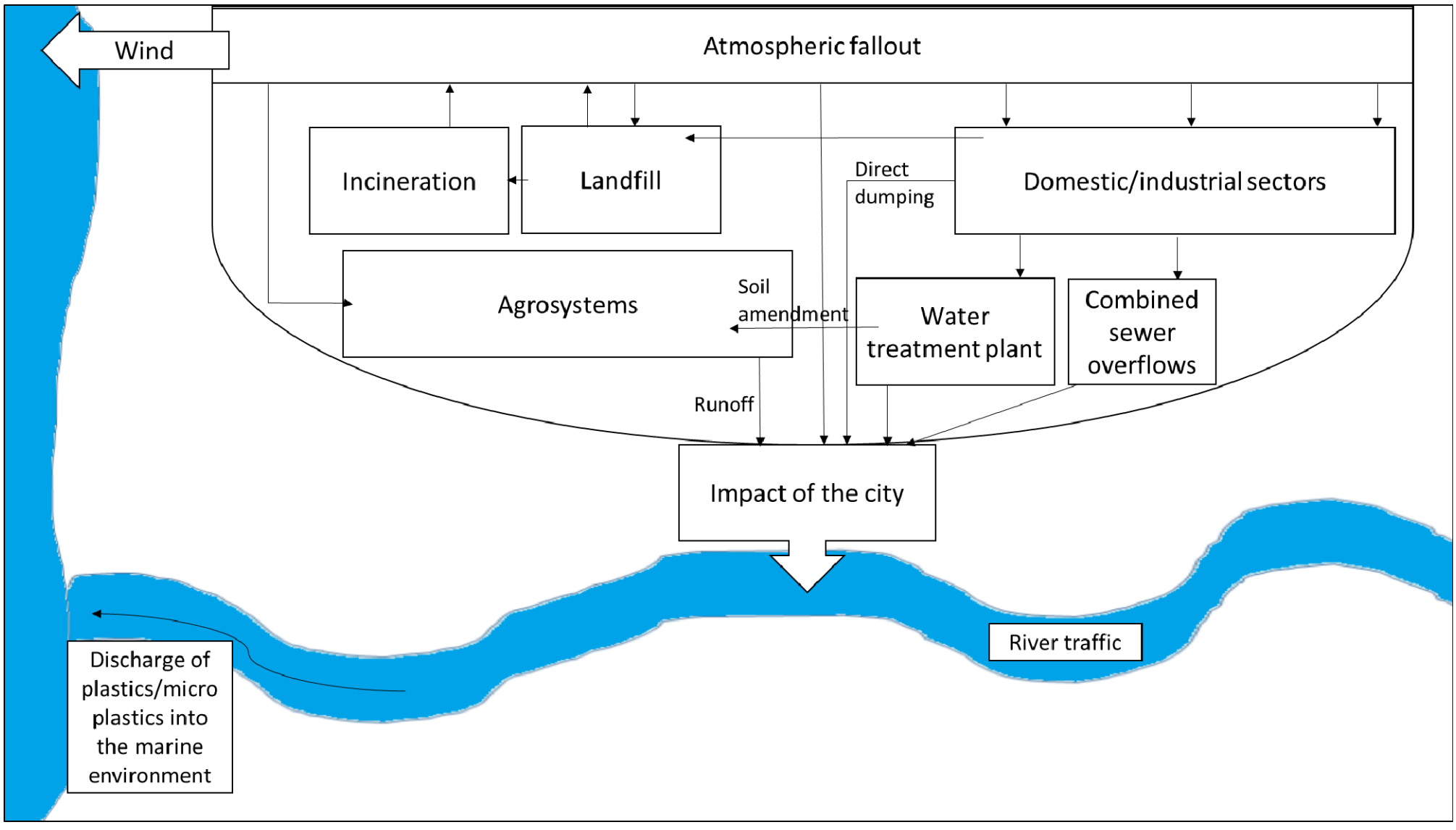
Figure 1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Incineration
Landfill
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Domestic/industrial sectors
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Runoff
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River traffic
Discharge of
plastics/micro
plastics into
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Figure 2

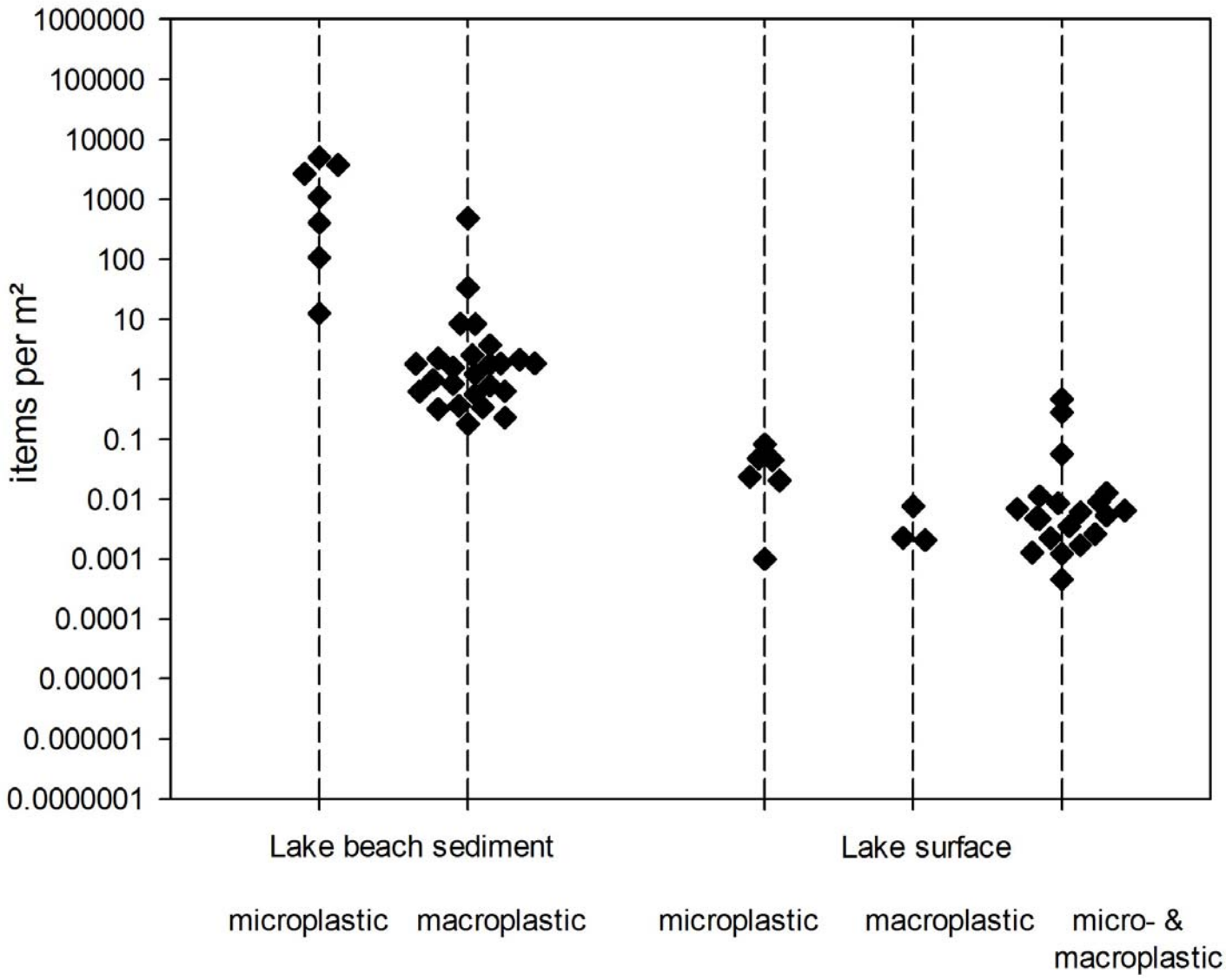
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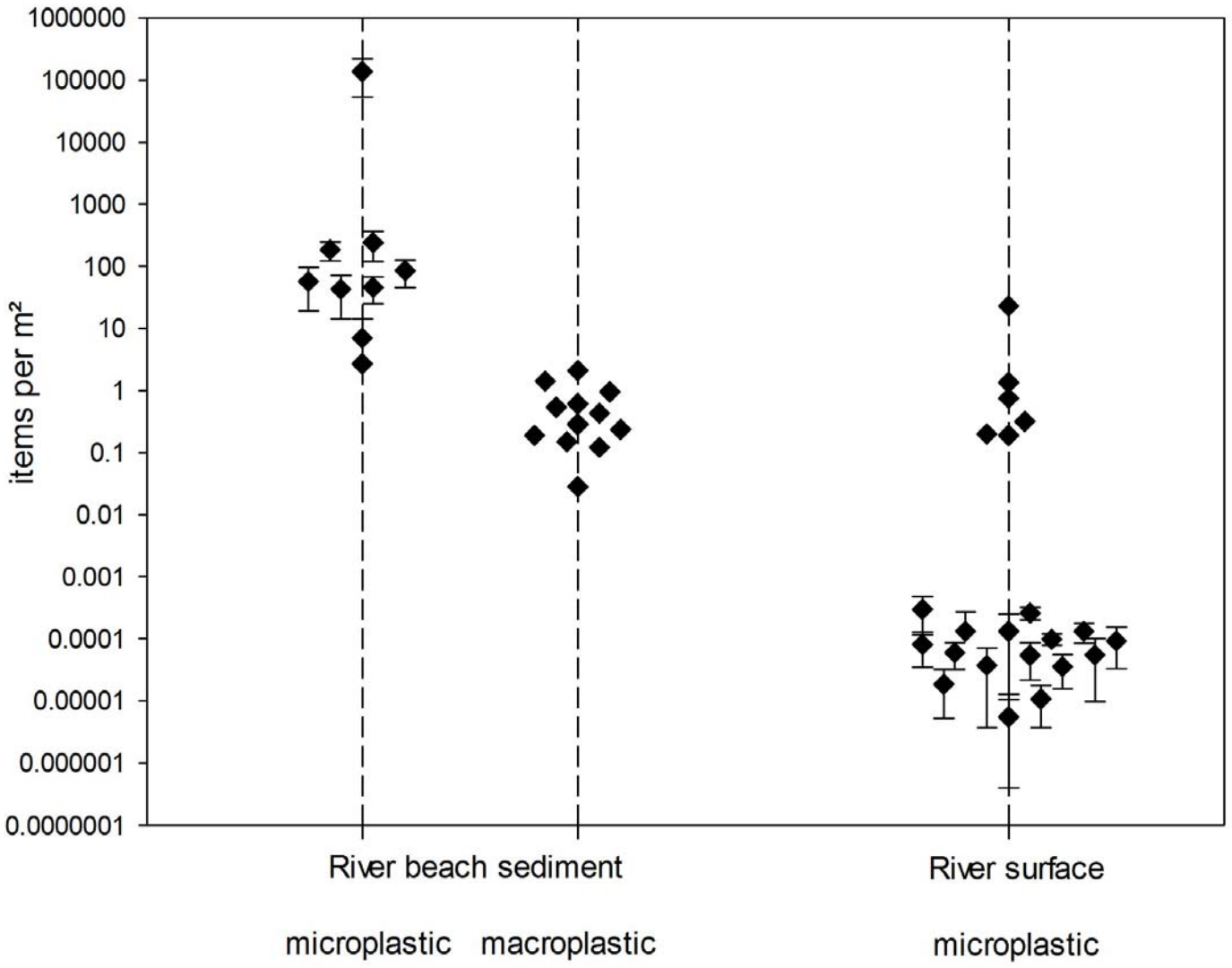
Figure 3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items per m²
River beach sediment
microplastic
macroplastic
River surface
microplastic

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Table 1

Environment	Year	Site	Compartment(s) studied	Sampling methods	Identification-method	Unit used	Reference
Lakes	2011*	Lake Huron (Canada, USA)	Lakeshore sediments	Visual inspection and plastic particles	FT-IR	Particles/m^2	[10]
	2012	Lake Geneva (Switzerland, France)	Lakeshore sediments	Manual collection of sediment samples	Visual inspection	Number of particles in 1L samples	[28]
			Biota (fish and birds)	-		-	
			Lake water	Manta trawl		Particles/km^2	
	2013*	Lake Garda (Italy)	Lakeshore sediments	Random grid sediment sampling	RM	Particles/m^2	[9]
	2013*	Lakes Superior, Huron and Erie (USA, Canada)	Lake water	Manta trawl	SEM/EDS	Particles/km^2	[7]
	2013*	Lake Geneva (Switzerland, France)	Lakeshore sediments	Manual collection of sediment samples	Visual inspection	Particles/m^2	[29]
			Lake water	Manta trawl		Particles/km^2 and g/km^2	
	2014*	Lake Hovsgol (Mongolia)	Lake water	Manta trawl	Visual inspection	Particles/km^2	[35]
	2014*	Lake Erie and St. Clair (USA, Canada)	Shorelines	Manual collection of sediment samples	Randomly selected samples with FT-IR	Particles/m^2	[30]
Rivers	2010	Rivers in Greater London (UK)	River water	No information	IR-spectroscopy	Particles/L	[36]
	2011#	Los Angeles and San Gabriel Rivers (USA)	River water	Manta net Streambed samples Hand net (0.8, 0.5 mm mesh size)	Visual inspection	Particles/L	[37]
	2014#	Rivers Elqui, Maipo, Maule and BioBio (Chile)	Riversides sediments	Visual sampling (items >1.5cm)	Visual inspection	Number of items	[33]
	2014	Thames river (UK)	Subsurface water	Eel nets	Visual inspection	Numbers of items sampled	[41]
	2014	Danube River (Germany, Austria etc.)	River water	Stationary driftnets (0.5 mm mesh size)	Density separation & Visual inspection	Particles/1000 m^3	[38]
	2014	Seine River (France)	Floating plastics in river water	Floating booms	FTIR-ATR	Tons of plastic	[42]
	2014#	St. Lawrence River (Canada)	Bank sediments	Grab samplers	Differential scanning calorymetrie	Particles/m^2	[32]
	2014#	Four Estuarine Rivers in the Chesapeake Bay (USA)	River water	Manta Trawl	RM	g/km^2	[40]
	2015#	Seine & Marne River (France)	River water	Manta Trawl (330 µm) & Plankton net (80 µm)	Visual inspection	Particles/m^3	[22]

RM = Raman microspectroscopy